CHAPTER

3 Microbial enzymatic remediation of synthetic plastic pollutants: a present and future perspective

N. Prabhu[1], M. Rajamehala[2], S. Chozhavendhan[2] and T. Gajendran[3]

[1]*Sethu Institute of Technology (Autonomous), Kariapatti, Tamil Nadu, India,*

[2]*Department of Biotechnology, Vivekanandha College of Engineering for Women (Autonomous), Namakkal, Tamil Nadu, India,*

[3]*Department of Biotechnology University College of Engineering, BIT Campus, Anna University, Tiruchirappalli, Tamil Nadu, India*

3.1 Introduction

Plastics primarily consist of synthetic polymers composed of carbon atoms linked together with hydrogen, oxygen, nitrogen, and sulfur, forming a framework around the carbon chains (Shah et al., 2008). Polymers have become ubiquitous in various aspects of human existence since their inception in the 1950s. The manufacturing of plastics has significantly increased over time to cater to the daily needs of the global population. According to Kamboj (2016), the production of plastic over the past 70 years has reached approximately 8.3 billion tonnes. The significant rise in plastic production is mostly attributed to the fabrication of single-use disposable plastic materials, which constitute approximately 50% of the total plastic products. The disposal of plastics has significantly increased, resulting in contamination of both land and oceans. Plastics are extremely nonbiodegradable, leading to their accumulation in the environment and subsequent environmental damage (Shimao, 2001). Approximately 21% of the global plastic production is effectively managed, while the other portion persists in the environment, either through terrestrial disposal, resulting in accumulation and environmental contamination, or through marine disposal, contributing to water pollution.

The presence of plastic garbage in the environment leads to chemical pollution as a result of employing disposal techniques such as incineration. Chemicals commonly utilized in consumer plastic products, such as ethylene, vinyl chloride, terephthalic acid (TPA), and bisphenol, have been found to pose detrimental effects on human health. These effects include the development of diseases such as cancer,

Plastic Waste Valorization. DOI: https://doi.org/10.1016/B978-0-443-41630-9.00017-3
© 2026 Elsevier Inc. All rights are reserved, including those for text and data mining, AI training, and similar technologies.

endocrine disorders, birth defects, and various other health complications (Waring et al., 2018).

There is a growing interest among individuals to explore more effective and expeditious methods of managing the extensive accumulation of this hazardous substance, given the inherent challenges associated with entirely discontinuing the manufacturing of these very adaptable and widely utilized plastic commodities. The solution likely resides within the purview of our scientific community, which is diligently engaged in developing a biologically sound method for effectively managing the escalating accumulation of plastic garbage (Trevino et al., 2012). Tsushima and Matsushita (2010) have identified specific microorganisms that possess the ability to manufacture enzymes capable of inducing degradation of such polymers. The enzymes under consideration are classified within the hydrolase family, encompassing esterases, lipases, depolymerases, and PETases. These enzymes have been identified for their ability to degrade the carbon backbone of many widely utilized plastics (Pometto et al., 1992). These enzymes exhibit their activity in the presence of water, thus earning the designation of hydrolytic enzymes. They catalyze the breakdown of the plastic polymer into simpler monomeric units, facilitating their assimilation by microorganisms as a carbon source. Subsequently, these monomeric units are further metabolized into CO_2, H_2O, CH_4, and N_2 (Seo et al., 2009). The utilization of metabolic byproducts from the degradation of plastics by microorganisms holds significant potential for addressing the pressing issue of plastic pollution. By repurposing these byproducts in various applications, a sustainable recycling cycle can be established, effectively mitigating the environmental harm caused by these persistent materials (Villain et al., 1995). This study provides a contemporary survey of the research conducted in this field, focusing on the ongoing worry regarding the biodegradability rate of plastics, which remains a significant global issue.

Plastic-degrading enzymes, similar to other enzymes engaged in the biological breakdown of polymers, have been categorized into two main groups: extracellular enzymes and intracellular enzymes (Gu, 2003). Nevertheless, the category that has received the most extensive research attention is the extracellular enzymes. These enzymes exhibit a diverse array of reactivity, encompassing both oxidative and hydrolytic functionalities (Glaser, 2019). The primary function of these entities is to engage in the depolymerization process of the extended carbon chains found within plastic polymers, resulting in the formation of a combination of oligomers, dimers, and occasionally, monomers. The aforementioned enzymes, which exhibit similar characteristics to microbial laccases, peroxidases, lipases, esterases, and cutinases, have been identified as belonging to these respective enzyme classes (Gan and Zhang, 2019). Moreover, it has been suggested that these extracellular enzymes play a role in diverse events that take place at the interface between a solid and liquid phase. Specifically, they catalyze reactions on the macromolecules present on the surface of solid plastic when immersed in a liquid phase (Chinaglia et al., 2018). Additional enzyme groups participate in the process of modifying the hydrophobic plastic surfaces, breaking down plastic metabolic intermediates into individual

monomeric units, and ultimately converting the resulting monomeric intermediates into minerals. A significant fraction of intracellular enzymes play a crucial role in facilitating the aerobic and anaerobic processes required for the conversion of intermediates into chemicals that can be ingested by microorganisms (Pathak and Navneet, 2017). In this chapter, the plastic-degrading enzymes are categorized based on the specific polymer they target, namely, polyethylene (PE), polyurethane (PU), polyethylene terephthalate (PET), polystyrene (PS), and nylon.

3.2 Types of plastics frequently used

Plastics have significantly transformed several industries as a result of their inherent versatility and the ability to manipulate this polymer via the use of diverse chemical compounds (Vona et al., 1965). These objects are designed with distinct characteristics that are tailored for specific uses (Pavia et al., 1988). The next section provides a description of some frequently utilized plastics for diverse applications, along with an overview of their primary uses across various industries.

3.2.1 Polyethylene

PE is one of the most widely used plastic types. It is a member of the thermoplastics class and is a polymer of ethylene monomers. Over the years, millions of tons have been manufactured, and since its commercial manufacturing started in the 1950s, it has been utilized in numerous sectors and fields. High-density PE is employed in the construction industry, whereas low-density PE is used in carry bags. These two types of PE are used in quite different contexts.

3.2.2 Polyethylene terephthalate

The most popular synthetic polymer utilized in the production of single-use, throwaway beverage bottles is terephthalate, which is incredibly robust and hard. This thermoplastic material is created by creating ester bonds during the polymerization of terephthalic and ethylene acid. One of the primary causes of plastic pollution is the single-use plastic products that are discarded on land or in the water. The toughness and strength of PET repeating units are attributed to their massive aromatic ring (Gross and Kalra, 2002).

3.2.3 Polypropylene

One of the most widely used plastics, polypropylene (PP), is made of propylene monomers and was first polymerized in 1951 (Carniel et al., 2016). This thermoplastic has been highly recognized in a variety of industries, including the

textile, automotive, and packaging sectors (Williams, 1981). This plastic's ability to copolymerize with other polymers, expanding the diversity of products that can be made accessible according to various needs, is one of its main advantages (Rowe et al., 2002). Being a thermoplastic, PP can take on various shapes when heated up without losing its structural integrity. As a result, it has numerous industrial uses where it is quite helpful (Nakkabi et al., 2015).

3.2.4 Polyvinyl chloride

Polyvinyl chloride (PVC) is the third most popular synthetic plastic substance worldwide, behind PE and PP. Before the plasticizers are added, the material appears as a brittle white substance. There are two main forms that it can be made in: stiff and flexible. Rigid PVC is given mechanical qualities that are more flexible and smooth by adding plasticizers, such as phthalates. PVC is generally much harder, more affordable, and has a far higher tensile strength than other plastics on the market. Nevertheless, burning it releases hydrogen chloride vapors, which can be harmful to the health of people exposed to them. For this reason, it is not advised in devices such as electrical wire insulators (Vona et al., 1965).

3.2.5 Polystyrene

A homopolymer made of styrene repeating units is called PS. Depending on its type, it can be either a thermoplastic or a thermosetting plastic. It is a transparent thermoplastic substance that exists naturally. This plastic is a pioneer in the packaging sector because its stiff form, a sort of foam material, is utilized in most applications. Because PS has no smell and is nontoxic, it is a great material for food packing. However, because it is tossed and disposed of primarily, it is relatively inert and offers a significant environmental contamination risk (Tokiwa et al., 2009).

3.2.6 Polylactic acid

Since polylactic acid (PLA) plastic is made from renewable resources like sugar cane and maize starch, it differs from many other plastics. For this reason, it is called a bioplastic. It is biodegradable and shares characteristics with PE, PP, and PS (Gross and Kalra, 2002). Compared with other plastics that take over 500 years to disintegrate, natural breakdown usually occurs in a little as 6 weeks to 24 months. It is among the safest plastics to use globally due to its cost-effective manufacture (Williams, 1981).

3.2.7 Polybutylene succinate

The thermoplastic polybutylene succinate (PBS) has characteristics similar to those of PP. Because it naturally breaks down into carbon dioxide and water, it is a

biodegradable plastic that can be utilized successfully. Succinic acid and 1,4-butanediol are used in its synthesis. It requires no plasticizers to produce, making it more pliable than PLA polymers (Gross and Kalra, 2002).

3.2.8 Polyurethane

Organic units connected by carbamate linkages are used to make PU. They could be ether or an ester. They do not dissolve in water. As one of the most versatile types of plastic, it is used to make sealants, elastomers, medical devices, flexible and rigid foams, and a variety of other products (Nakkabi et al., 2015).

3.3 Enzymatic degradation of plastic

Biocatalysts known as enzymes take part in a reaction, operate on a specific substrate, and quicken the process of turning that substrate into a useful product. There is a growing interest in the fundamental mechanism by which microbes can break down this extremely resilient polymer due to the possibility of microorganisms degrading plastic that pollutes our environment. As previously mentioned, the primary degradation is facilitated by extracellular and intracellular microbial enzymes, which break down polymer chains by assimilation within the cell and release metabolic products such as CO_2, H_2O, CH_4, and N_2 (Amobonye et al., 2021).

Numerous enzymes that are known to degrade various types of plastic materials have been identified from bacteria, fungi, actinomycetes, and algae (Urbanek et al., 2020). The class "Hydrolases" includes all known enzymes that break down polymeric polymers. These enzymes participate in a catalytic process that, in the presence of water, breaks down the chemical bonds in their substrate (Tokiwa et al., 1976). Recent studies in this field have identified cutinase, lipase, and PETase (an esterase) as some of the most prevalent enzymes linked to the breakdown of plastics (Tokiwa and Suzuki, 1977a, 1977b). Similar to each other, these enzymes work on the plastic polymer by hydrolytically breaking the long carbon chains into smaller subunits, which are subsequently assimilated into the microbial cell for additional enzymatic breakdown and the release of metabolic products (Tokiwa and Suzuki, 1977a, 1977b). One of the primary study areas to address the ever-increasing plastic pollution in the environment is the extraction of these enzymes and their modification to enhance their enzymatic activity. It has been reported that in the presence of these enzymes, a variety of plastic types, including PE, PET, PLA, PBS, and PU, degrade slowly (Mukai et al., 1993). The next section discusses the mechanism of these polymers' enzymatic breakdown.

3.4 Mechanism of enzymatic degradation

The primary enzymes involved in the breakdown of environmental plastic compounds are hydrolases (Müller et al., 2005). According to Müller et al. (2006), the third class of enzymes is represented by the enzyme class "Hydrolases." These are responsible for the cleavage of chemical bonds in the presence of water, which breaks bigger molecules into smaller ones. The following equation represents the general response of a hydrolase enzyme to the breakdown of a product (A-B) in the presence of water:

$$A\text{-}B + H_2O \rightarrow A\text{-}OH + B\text{–}H$$

The two-step process of cleaving lengthy carbon chains is caused by the hydrolases that break down polymeric polymers. Every plastic found in the environment is hydrophobic by nature. In the first step of the enzyme-polymer interaction, the extracellular enzymes produced by different microorganisms attach themselves to the plastic surface through hydrophobic interactions. Many hydrolases have a hydrophobic cleft at the active site that can accept the hydrophobic groups in the polymer, improving the enzyme's accessibility to the polymer (Wilkes and Aristilde, 2017). The lengthy polymer chains are hydrolytically broken into smaller monomers or dimers in the second stage of the reaction by the enzyme's active site. The microbial organism can then concentrate and use these smaller monomers or dimers as a source of carbon (Barth et al., 2015).

3.5 Factors affecting plastic degradation

The breakdown of synthetic plastics is influenced by the characteristics of polymers and their sensitivity to various biotic and abiotic stressors (Artham and Doble, 2008). The breakdown of a substance is influenced by its physicochemical qualities, namely, its vulnerability to degradation by biotic or abiotic factors, which is governed by the composition of its backbone and the length of its chain. Polymeric materials are characterized by the presence of extended carbon chains, as shown in PP, which confer resistance to biodegradation. Nevertheless, the incorporation of heteroatoms into the carbon chain, such as in the context of polymers containing oxygen, makes them susceptible to thermal breakdown and biodegradation. Moreover, the efficiency of polymer breakdown is influenced by hydrophobicity as an additional component. The degradation rate increases with an increase in hydrophilicity (Ali et al., 2021).

The degree of crystallinity has a significant influence on the rate at which plastic materials undergo deterioration. A higher degree of polymer crystallinity necessitates an increased demand for oxygen and water, potentially initiating the breakdown process. The degradation process is influenced by the amorphous regions of polymers, which are believed to exhibit greater sensitivity to thermal oxidation. In addition, the degradation rate of a polymer may be influenced by its molecular weight. According to

this concept, polymers of a high molecular weight exhibit a slower degradation rate because to their comparatively reduced surface area (Kour et al., 2023).

The process of plastic polymer manufacture incorporates the utilization of additives, which have the potential to influence the rate of deterioration. Nanoadditives have the potential to enhance the polymeric characteristics, hence offering significant improvements in various industrial applications. The utilization of nanoscale reinforcements leads to an augmentation in the surface area of the interface, hence resulting in enhanced performance. The incorporation of these nanoparticles into the polymers aims to enhance their mechanical, rheological, electrical, and thermal characteristics. Furthermore, this facilitates the recycling and degradation process. The incorporation of nanoparticles into polymer manufacturing processes has the potential to address challenges related to biodegradation (de Dicastillo et al., 2020). The degradability of the polymer may be affected by the manufacturing process. Copolymerized PP, in comparison with PP produced through bulk polymerization or employing the Ziegler-Natta catalyst, exhibits reduced susceptibility to photodegradation.

Stabilizers are commonly utilized as additives in the production of plastics in order to mitigate the rate of degradation. The process of chemical functionalization inside a polymeric structure results in permanent alterations to its physicochemical properties and degradation rate. The degradability of the polymer is influenced by its morphological features. The rate of degradation is shown to increase with an increase in surface roughness, since a bigger surface area facilitates the establishment of biofilms to a greater extent compared with smooth surfaces (Booth et al., 2017). The mechanisms and rate of plastic degradation may be influenced by factors such as geographical location, levels of air pollution, and prevailing weather conditions. As an illustration, it has been shown that PET bottles have the potential to persist on the seabed for a duration exceeding 15 years (Fotopoulou and Karapanagioti, 2017). The degradation process is significantly influenced by the availability of water, as the hydrolytic cleavage of susceptible functional groups leads to the separation of polymeric chains (Pitt, 1992). The decomposition rate of plastics is determined by the availability of oxygen. The presence of abundant oxygen levels results in an expedited process of polymeric breakdown as a consequence of the reactivity between oxygen and carbon-centered radicals that are formed in the initial stages of degradation (Price and Horrocks, 2013).

3.6 Recent advances in plastic degradation by enzymes

The enzymes that have been the subject of recent research in the enzymatic breakdown of plastics primarily include esterase, cutinase, lipase, and PETases (Kyrikou and Briassoulis, 2007). The aforementioned enzymes play a role in the degradation of several types of plastic polymers, as detailed by Yamashita et al. (2019) (Table 3.1).

Table 3.1 Microbial enzymes involved in the degradation of different types of plastics.

Plastic degrading microbes	Plastic used	Enzyme	References
Acidovorax delafieldii	Poly(tetramethylene succinate)-*co*-adipate	Lipase	Uchida et al. (2000)
Acinetobacter gerneri	Polyurethane	Polyurethanase	Howard et al. (2012)
Acinetobacter lwoffii	Polyhydroxyalkanoates	Lipase	Sharma et al. (2019)
Actinomadura sp.	Polyester	Depolymerases	Sriyapai et al. (2018)
Alcaligenes faecalis	Polyethylene	Lipase	Nag et al. (2021)
Alcaligenes faecalis	Polyethylene	CMCase	Nag et al. (2021)
Alcaligenes faecalis	Polyethylene	Xylanases	Nag et al. (2021)
Alcaligenes faecalis	Polyethylene	Protease	Nag et al. (2021)
Amycolatopsis mediterannei	Poly(ε-caprolactone)	Cutinase	Tan et al. (2021)
Aspergillus melleus	Poly(ε-caprolactone)	Lipase	Amin et al. (2022)
Aspergillus niger	Poly(lacticacid)	Lipase	Nakajima-Kambe et al. (2012)
Aspergillus niger	Poly(ε-caprolactone)	Lipase	Nakajima-Kambe et al. (2012)
Aspergillus tamarii	Polyethylene terephthalate	Lipase	Anbalagan et al. (2021)
Aspergillus tamarii	Polyethylene terephthalate	Cutinase	Anbalagan et al. (2021)
Bacillus pumilus	Polyurethane	Lipase	Nair and Kumar (2007)
Bacillus sp.	Poly(butyleneadipate coterephthalate)	Lipase	Zhang et al. (2022)
Bacillus subtilis	Polyurethane	Polyurethanase-lipase	Rowe and Howard (2002)
Bacillus sp.	Cellulose acetate plastic	Lipase	Ishigaki et al. (2000)
Bacillus sp.	Cellulose acetate plastic	Cellulase	Ishigaki et al. (2000)
Candida antarctica	Poly(butylenesuccinate)	Cutinase	Shi et al. (2019)
Fusarium sp.	Poly(butylenesuccinate)	Lipase	Shi et al. (2019)
Ideonella sakaiensis	Polyethyleneterephthalate	PETase	Alfieri et al. (2022)
Laceyella sp.	Polyester	Depolymerases	Sriyapai et al. (2018)
Lactobacillus brevis	Poly(ε-caprolactone)	Lipase	Khan et al., 2017
Lactobacillus plantarum	Poly(ε-caprolactone)	Lipase	Khan et al., 2017
Moraxella sp.	Synthetic polymers	Esterase	Nikolaivits et al. (2022)
Penicillium crustosum	Polyethylene terephthalate	Lipase	Anbalagan et al. (2021)
Penicillium crustosum	Polyethylene terephthalate	Cutinase	Anbalagan et al. (2021)

Pseudomonas chlororaphis	Polyhydroxy alkanoates	Lipase	Sharma et al. (2019)
Pseudomonas chlororaphis	–	Lipase	Mohanan et al. (2022)
Saccharothrix sp.	Poly(butylenes succinate)	Depolymerases	Sriyapai et al. (2019)
Stenotrophomonas sp.	Poly(butylenes adipate-*co*-terephthalate)	Lipase	Jia et al. (2021)
Streptomyces sp.	Polyester	Depolymerases	Sriyapai et al. (2018)
Thermobifida alba	Aliphatic-aromatic co poly ester film	Esterase	Hu et al. (2010)
Pseudozyma antarctica	Biodegradable plastic	Esterase	Yamashita et al. (2019)
Cryptococcus sp. strain S-2	PLA	Cutinase like enzyme	Masaki et al. (2005)
Cryptococcus sp.	PBS	Lipase	Thirunavukarasu et al. (2016)
Aspergillus fumigatus	PHB, PBS, and PES	Poly hydroxy butyrate depolymerase	Thirunavukarasu et al. (2008), Jung et al. (2018), Kasuya et al. (1998)
–	PCL	Bifunctional lipase-cutinase	Liu et al. (2019)
Fusarium solani	PBS	Cutinase	Hu et al. (2016)
Pichia pastoris	PBS	Cutinase	
Candida rugosa	PBS	Lipase	Pereira et al. (2001)
Ideonella sakaiensis	PET	PETase	Webb et al. (2013), Paci and La Mantia (1999)
Humicola insolens and *Thermobifida cellulosilytica*	PBTF and PBF	Cutinase	Cardi et al. (Giglia et al., 2019)
Candida rugosa/Tritirachium album	PLA/cellulose nanocrystal composites	Lipase/proteinase K	Hegyesi et al. (2019)
Ideonella sakaiensis	PETase and MHETase	PET	Knott et al. (2020)

PBF, Poly(1,4-butylene 2,5-furandicarboxylate); ***PBS***, polybutylene succinate; ***PBTF***, poly(1,4-butylene 2,5-thiophenedicarboxylate); ***PCL***, poly Ɛ-caprolactone; ***PES***, polyethylene succinate; ***PET***, polyethylene terephthalate; ***PHB***, polyhydroxybutyrate; ***PLA***, polylactic acid.

3.6.1 Cutinases

Cutinases are a specific type of esterase enzyme that may be distinguished by their capacity to catalyze the hydrolysis of polyesters with a significant molar mass (Chen et al., 2013). The enzymes in question are classified as α/β hydrolases or carboxylic ester hydrolases. These enzymes have been identified in plant pathogenic fungi, specifically *Fusarium solani pisi*, as documented by Kolattukudy and Brown (1975), Kolattukudy et al. (1981), and Heredia (2003). Cutinases are synthesized by several fungal species such as *Fusarium solani* (Alisch-Mark et al., 2006; O'Neill et al., 2007), *Penicillium citrinum* (Liebminger et al., 2007), *Pichia pastoris* (Munari, 2019), *Aspergillus oryzae*, and *Humicola insolens*. The study conducted by Ronqvist et al. (2009) demonstrated that the enzymatic actions of *Fusarium solani pisi* cutinase and *Humicola insolens* cutinase (HiC) exhibited the ability to degrade low crystallinity PET film. The results indicated a substantial weight loss of 97% within a time frame of 96 hours. PET-hydrolases are a subset of enzymes known as cutinases, which have demonstrated considerable potential in the process of PET biodegradation. An enzyme with comparable functionality with cutinase, specifically obtained from *Cryptococcus* sp., The strain S-2 was characterized for its efficient breakdown of high–molecular-weight plastic, specifically PLA-based plastic, as reported by Van Gemeren et al. in 1998. The study conducted by Furukawa et al. (2019) demonstrated the enzymatic degradation of electrospun polycaprolactone fiber mats by Cutinase 2p derived from *Arxula adeninivorans*.

The expression of cutinase from *Fusarium solani* in *Pichia pastoris* has been employed to achieve the enhanced production of the enzyme, to facilitate the degradation of PBS plastic (Hu et al., 2016). According to Marija and Jasna (2001), the recombinant cutinase showed a notable capacity to expedite the degradation process. Remarkably, within a just 6 hours, the PBS films underwent total breakdown. The presence of perforations on the PBS films provided evidence that the enzyme's impact was optimized with longer degrading periods (Gan et al., 2001). The investigation conducted by Maeda et al. (2005) demonstrated the characterization of PBS films that had undergone enzymatic degradation. The results indicated that both the amorphous and crystalline structures of PBS were affected by the recombinant enzyme. In a separate study investigating the degradation of PBS films with varying molecular weights, it was hypothesized that films with larger molecular weights had a greater propensity for accelerated and facilitated disintegration (Pan et al., 2018). Previous studies have demonstrated the ability of cutinases to effectively decompose several forms of biodegradable plastics (Masaki et al., 2005). van Gemeren et al. (1998) reported the discovery of an enzyme with functional similarities to cutinase, derived from yeast of the *Cryptococcus* sp. strain S-2. This enzyme exhibited the capability to degrade a high–molecular-weight plastic substance composed of PLA. The enzyme demonstrated the capability to degrade PLA in a time frame of 60 hours, using a minimal enzyme concentration of 0.8 μg mL^{-1}. This degradation rate is notably faster when

compared with other enzymes documented for their ability to dissolve PLA plastic (Oda et al., 2000).

3.6.2 Lipases

Lipases are a type of enzyme that facilitates the breakdown of lipids. They belong to the subclass of esterase enzymes. Several species, such as *Rhizopus delemer*, *Candida antarctica*, *Thermomyces lanuginosus*, and *Candida rugosa*, have gained recognition for their ability to produce lipases and participate in the degradation of plastics. For instance, Vertommen et al. (2005) reported the involvement of *Candida antarctica* in this process, while Eberl et al. (2009) observed the contribution of *Thermomyces lanuginosus*. These species were found to be capable of degrading poly (butylene succinate-*co*-hexamethylene succinate) copolymer. In a study conducted by Tokiwa and Calabia (2009), it was observed that the lipase produced by *Rhizopus delemer* was able to break down around 53% of the polyester type-polyurethanes (ES-PU) film within a 24-hour reaction period (Pereira et al., 2001). In a study conducted by Thirunavukarasu et al. (2008), it was shown that a lipase enzyme derived from the yeast *Cryptococcus sp.* demonstrated the ability to hydrolyze PBS and polybutylene succinate-*co*-adipate (PBSA). The hydrolysis of PET to TPA by Lipase B derived from *Candida antarctica* was observed to be effective (Carniel et al., 2017). Additionally, the enzymatic decomposition of electrospun polycaprolactone fiber was demonstrated by Lipase FE-01 obtained from *Thermomyces lanuginosus* (Furukawa et al., 2019).

The study conducted by Thirunavukarasu et al. (2016) revealed that a total breakdown of plastic materials occurred within a timeframe of 72 hours for PBS and 16 hours for PBSA. The authors of the study conducted Fourier transform infrared spectroscopy analysis on the PBS and PBSA polymers that had undergone enzymatic degradation. The purpose of this analysis was to identify the specific chemical groups that were destroyed by the lipase enzyme, as reported by Kijchavengkul et al. (2010). A notable reduction in the transmittance of both the C–O bond and –C–O–C– bonds was detected when compared with the nondegraded film of PBS. Umare et al. (2007) demonstrated the occurrence of primary chain scission at the ester linkage, as evidenced by a large peak observed at 3372 cm^{-1}, which is indicative of the stretching vibration of the hydroxyl (–OH) group. In a study conducted by Rizzarelli and Carroccio (2009), it was shown that the peaks at 1041 cm^{-1} and 1332 cm^{-1}, which were observed in nondegraded polybutylene succinate adipate (PBSA), were not present in the hydrolytic product of the polymeric material. The observed peaks can be attributed to the stretching vibrations of the O–C–O and C–H bonds, respectively. The nuclear magnetic resonance study of the polymer films made from PBS and PBSA revealed that the enzymatic degradation resulted in the disappearance of the 1,4-butanediol signal. This absence suggests that hydrolysis of the carboxyl end of the polymer chain may have occurred (Li et al., 2011).

A lipase derived from *Candida rugosa* was employed for the purpose of degrading a copolymer composed of poly(butylene succinate-*co*-hexamethylene succinate). The research study posited that an increase in the high-styrene (HS) content of a copolymer corresponds to a heightened vulnerability to enzymatic degradation by lipase. The degradation of the P (BS-*co*-HS) copolymer was assessed by measuring weight loss, as described by Pereira et al. (2001).

The enzymes lipase and cutinase are classified as esterases, which are a subset of the hydrolases enzyme class. In a recent study conducted by Liu et al. (2019), it was demonstrated that a combination of these two enzymes exhibited efficacy in the enzymatic degradation of poly Ɛ-caprolactone (PCL), a type of plastic material. The construction of the enzyme fusion was achieved using the end-to-end fusion technique, and afterwards, the fusion system was over expressed in *Pichia pastoris*, as described by Aris et al. (2016). According to Khan et al. (2017), the breakdown rates of PCL with Lip-Cut fusion enzyme over 6 hours were significantly greater compared with the degradation rates seen when using the two enzymes separately. The utilization of gas chromatography-mass spectrometry technique facilitated the examination of the deteriorated substances. The findings indicated that the primary byproducts resulting from the degradation of polycaprolactone (PCL) were 6-hydroxyhexanoic acid and a little proportion of Ɛ-caprolactone (Shah et al., 2013).

3.6.3 Proteases

Proteases, which are a type of enzyme, facilitate the cleavage of lengthy peptide chains into shorter peptides or the breakdown of proteins into polypeptide chains through the process of hydrolysis. This process is referred to as proteolysis. The fungal species mentioned in the study conducted by Loredo-Treviño et al. (2011) and Cosgrove et al. (2007) include *Aspergillus*, *Trichoderma*, *Paecilomyces*, *Penicillium*, *Alternaria*, and *Fusarium*. Additionally, *Phaenarochaete* was identified as a relevant fungal species, while Russell et al. (2011) studied *Pestalotiopsis*. *Rhizopus, Mucor, Humicola, Thermoascus*, and *Thermomyces* represent a selection of significant fungal species that have been identified as producers of proteases for the purpose of plastic degradation. The research conducted by Ozsagiroglu et al. (2012) demonstrates that *Arthrobotrys oligospora* is capable of synthesizing a serine protease enzyme that has the ability to break down PLA.

3.6.4 Esterases

Esterases are a class of hydrolase enzymes that catalyze the hydrolysis of ester bonds, resulting in the formation of alcohols and acids through the addition of water molecules. Esterases play a significant role in the decomposition of plastics, a process that is carried out by both bacteria and fungi. Specifically, the esterase derived from *Comamonas acidovorans* is beneficial in the degradation of low–-molecular-weight PLA, a type of plastic derived from renewable resources.

Purpureocillium lilacinum and *Curvularia senegalensis* are two species of fungus known for their ability to degrade poly(butylene succinate-*co*-adipate) and PU materials, as reported by Yamamoto-Tamura et al. (2015). It is widely believed that *Aspergillus flavus* and *Aspergillus tubingensis* can release esterases, which play a crucial role in the degrading process of plastics (Khan et al., 2017; Tokiwa et al., 2009). The degradation of PU has been reported by Brunner et al. (2018) by the use of esterase, which is produced from *Xepiculopsis graminea* and *Penicillium griseofulvum*. According to Ganesh et al. (2017), lipase and esterase enzymes are capable of breaking down polycaprolactone polymers.

The acceleration of plastic mulch film disintegration in laboratory circumstances has recently been demonstrated by the esterase enzyme derived from the yeast *Pseudozyma antarctica*. The study conducted by Closas et al. (2016) showed that mulch films composed of biodegradable plastic had a significant breakdown rate when exposed to xylose during the growth of the strain. Following a 12-week incubation period of the plastic films with the enzyme, the scanning electron microscopy (SEM) investigation demonstrated the presence of pierced holes on the previously smooth surface of the films, accompanied by the emergence of an uneven surface. Furthermore, it was observed in the study conducted by Barragán et al. (2016) that the degradation of the amorphous part of the films occurred at a higher rate compared with the crystalline region.

3.6.5 Laccase

Laccases are a class of multicopper oxidases that facilitate the process of oxidizing phenolic chemicals. The process involves the utilization of molecular oxygen as a cosubstrate, resulting in the production of water and byproducts (Nunes and Kunamneni, 2018). The laccases possess a unique capability wherein they facilitate the oxidation of lignin, hence participating in the process of lignin degradation (Osma et al., 2010). Laccase has been found to participate in the oxidation process of the hydrocarbon backbone of PE, as documented by Sivan (2011). *Cochliobolus* sp., a particular fungal species, has been found to degrade PVC by the action of laccases, as reported by Srikanth et al. (2022). The strain TBB-03 of *Bjerkandera adusta* was recognized for its capacity to breakdown high-density PE by the synthesis of laccase, when subjected to treatment with lignocellulosic substrates (Bo et al., 2019). According to Osma et al. (2010), the fungi *Trametes versicolor* and *Pleurotus ostreatus*, as well as the bacteria *Streptomyces*, have been found to possess the enzyme laccase, which can degrade PE.

The ligninolytic enzyme groups typically consist of phenol oxidase (laccase), heme peroxidases, lignin peroxidase (LiP), manganese peroxidase (MnP), and versatile peroxidase (VP). Dashtban et al. (2010) and Phua et al. (1987) discovered that the degradation of medical polyester PU can be attributed to the hydrolysis of urethane and urea linkages, facilitated by the proteolytic enzymes papain and urease. This enzymatic process results in the production of free amine and hydroxyl

groups. The involvement of laccase produced by *Penicillium* in the breakdown of PE has been suggested by Abd El-Rehim et al. (2004).

3.6.6 Peroxidases

Peroxidases are a type of enzyme classified as oxidoreductases, responsible for facilitating oxidation-reduction reactions through the interaction of free radicals with molecules, resulting in the formation of oxidized and polymerized compounds. The enzymes known as catalases, which are also referred to as peroxidases in the context of fungi, are widely recognized for their superior efficacy in the conversion of lignin. The class under consideration encompasses LiPs, MnPs, and VPs, which are predominantly observed in white-rot fungi (Hofrichter and Ullrich, 2006). The study conducted by Iiyoshi et al. (1998) demonstrated the successful breakdown of high–molecular-weight PE by *Phanerochaete chrysosporium* and *Trametes versicolor*. The primary enzyme responsible for this degradation process was identified as MnPs. *Phanerochaete chrysosporium*, *Pleurotus ostreatus,* and S22 showed a notable proficiency in the degradation of lignin within the pH range of 9.0–11. The group of enzymes known as ligninolytic enzymes, which consists of LiP, MnP, and laccase, has been discussed in a study by Wu et al. (2005). According to Ganesh et al. (2017), the fungus *Fusarium graminearum* exhibited the ability to degrade PE through the production of peroxidase. The fungi *Aspergillus flavus*, *Aspergillus niger*, and *Fusarium graminearum* can create MnP and LiP enzymes. These enzymes play a significant role in the process of biodegradation of polyethylene carry bag. According to Bholay et al. (2012), *Aspergillus niger* and *Aspergillus flavus* have the highest range of LiP production.

3.7 Plastic-degrading enzyme classification based on type of polymer target

3.7.1 Polyethylene-degrading enzymes

Biocatalysts capable of degrading PE have been discovered in actinomycetal, bacterial, and fungal origins. These biocatalysts encompass a range of enzymes such as hydroxylases, laccases, peroxidases, and reductases. In a study conducted by Iiyoshi et al. (1998), it was shown that the breakdown of PE is primarily facilitated by a MnP enzyme. This enzyme was found to be prominently present in two lignin-degrading fungi species, namely, *Phanerochaete chrysosporium* and *Trametes versicolor*. In a study conducted by Yoon et al. (2012), it was shown that alkane hydroxylase, an enzyme, plays a significant role in the degradation of PE by *Pseudomonas* sp. E4. This finding was further supported by the cloning of the gene in *Escherichia coli* and subsequent assessment of the resulting recombinant protein. Jeon and Kim (2015) subsequently discovered that *P. aeruginosa* possesses an

enzyme system comprising alkane hydroxylase, rubredoxin, and rubredoxin reductase. This system was demonstrated to be accountable for the degradation of low–molecular-weight PE. Previous studies have demonstrated the noteworthy ability of laccases derived from actinomycetes, including *Rhodococcus ruber* (Santo et al., 2013), as well as fungi such as *Aspergillus flavus* and *Pleurotus ostreatus* (Gomez-Mendez et al., 2018), to effectively degrade PE. A proposal was put out suggesting that their action occurs through the oxidation of the PE hydrocarbon backbone.

3.7.2 Polyurethane-degrading enzymes

Various enzymes derived from bacteria and fungi, such as cutinases, esterases, lipases, laccases, peroxidases, proteases, and ureases, have demonstrated the capability to degrade PU (Magnin et al., 2020). The study conducted by Russell et al. (2011) emphasized the involvement of serine and cysteine hydrolase in the breakdown of Pestalotiopsis microspore PU. In a recent study conducted by Magnin et al. (2019), the researchers revealed the combined effects of esterase and amidase enzymes on the degradation of different PU derivatives. The study provided a stepwise process to explain the observed synergistic effects. The identification of enzymes involved in PU degradation within a microbial consortium was achieved by the application of proximity ligation-based metagenomic analysis. Gaytán et al. (2020) demonstrated that the enzymes were accountable for the metabolic processes of diverse PU intermediates. These enzymes encompassed a range of functional groups, such as dioxygenases, decarboxylases, dehydrogenases, transferases, ligases, hydrolases, isomerases, and peroxidases (Danso et al., 2019). It has been shown that the majority of PU-active enzymes investigated thus far have demonstrated activity towards ester-linked PUR, while exhibiting lower likelihood of activity against PU ethers. The presence of a carbonyl group in the ester-linked polyurethane (PUR) renders it more susceptible to hydrolysis induced by enzymatic processes, in contrast to the ether-linked PUR.

The process of PE biodegradation mediated by microbial enzymes can be divided into two distinct stages. The initial stage involves the binding of the enzyme to the PE substrate, which is subsequently followed by the process of hydrolytic cleavage. The biodegradation of PE is facilitated by intracellular and extracellular depolymerases that are synthesized by fungi and bacteria (Bhardwaj et al., 2013). Previous studies have documented the involvement of microbial enzymes in the process of lignin degradation, which has been found to have implications for the biodegradation of PE (Carrott and Carrott, 2007; Wei and Zimmermann, 2017; Restrepo-Flórez et al., 2014; Krueger et al., 2015). The enzymes encompassed in this category are laccases, LiPs, and MnPs. The involvement of MnP derived from *Phanerochaete chrysosporium* in the breakdown process of a PE membrane with a high molecular weight has been documented in a previous study (Iiyoshi et al., 1998).

The hydrolytic capacity of many enzymes has been demonstrated in emulsifying polyester PUR. These enzymes include the purified protease derived from *Pseudomonas fluorescens*, an esterase obtained from *Comamonas* acid*ovorans*, three esterases derived from *Pseudomonas chlororaphis*, and a lipase obtained from *Bacillus subtilis* (Rowe and Howard, 2002; Ruiz and Howard, 1999; Allen et al., 1999; Howard et al., 1999; Vega et al., 1999). The degradation of UV-irradiated films of PE has been studied using a thermostable laccase derived from *Rhodococcus ruber* (Santo et al., 2013).

3.7.3 Polyethylene terephthalate–degrading enzymes

Since the initial report by Müller et al. (2005) on the enzymatic hydrolysis of PET polymers using hydrolases from *Thermobifida fusca*, subsequent studies have identified several other enzymes, such as cutinases, esterases (Jabloune et al., 2020), lipases, and carboxylesterases, that possess the capability to degrade PET (Danso et al., 2019; Jabloune et al., 2020; Ru et al., 2020). According to Glaser (2019), the process of enzymatic hydrolysis of PET is predominantly characterized by a surface erosion mechanism. PETase, an aromatic polyesterase, derived from *I. sakaiensis* 201-F6, is well recognized as one of the most extensively investigated enzymes involved in plastic degradation. This enzyme has a basic α/β hydrolase shape. The enzyme in question facilitates the metabolic conversion of PET into bis(2-hydroxyethyl)-terephthalate, mono(2-hydroxyethyl)-terephthalate (MHET), and TPA. Additionally, an accompanying enzyme known as MHETase catalyzes the transformation of MHET into TPA and ethylene glycol (Austin et al., 2018). However, it should be noted that the enzyme exhibited thermolability. Consequently, efforts were made to enhance the thermostability of the PETase enzyme by engineering techniques. As a consequence, a novel variation was developed, which showed a 14-fold increase in activity compared with the original biocatalyst (Son et al., 2019). In a study conducted by Jabloune et al. (2020), a suberinase enzyme with a molecular weight of 25 kDa was identified from *Streptomyces scabies*. This particular suberinase exhibited exceptional stability and showed the ability to hydrolyze PET into TPA, along with other types of polymers.

PET plastic, commonly employed in the production of numerous throwaway plastic items, was previously believed to possess nonbiodegradable properties (Joo et al., 2018). In a recent study conducted by a group of researchers in Japan, a bacterium was discovered that exhibits growth capabilities on pet plastic trash that has accumulated over an extended period (Webb et al., 2013). The bacterial strain known as *Ideonella sakaiensis* was identified for its unique ability to metabolize PET waste bottles, utilizing PET as its primary carbon source for growth. This remarkable degradation process is facilitated by an enzyme called PETase, which enables the microorganism to break down PET plastic into valuable monomers, specifically TPA and ethylene glycol (Paci and La Mantia, 1999). The enzyme's active site demonstrated a high affinity for the synthetic polymer, allowing for facile

accommodation through hydrophobic contacts. Subsequently, the enzyme effectively initiated the process of hydrolytic degradation of PET plastic (Paci and La Mantia, 1998).

The carboxylesterases derived from *Thermobifida fusca*, *Bacillus subtilis*, and *Bacillus licheniformis* have been seen to exhibit partial hydrolysis of PET fibers, along with a notable capacity to degrade PET oligomers (Billig et al., 2010; Barth et al., 2016; Lülsdorf et al., 2015; Oeser et al., 2010; Ribitsch et al., 2011).

3.7.4 Polystyrene-degrading enzymes

Despite the existing evidence of microbial breakdown of PS by different bacteria and fungi, the specific enzymes responsible for the first depolymerization of these polymers have yet to be definitively identified. Nevertheless, a study conducted by Tahir et al. (2013) showed the ability of an extracellular esterase derived from *Lentinus tigrinus* to degrade PS. Furthermore, it has been observed that some polymerases derived from *Bacillus* and *Pseudomonas* species are also implicated in the breakdown of PS (Mohan et al., 2016). Several enzymes have been discovered as being involved in the metabolic process of styrene, the monomer of PS, before it entered into the tricarboxylic acid cycle. These enzymes include styrene monooxygenase, styrene oxide isomerase, phenylacetaldehyde dehydrogenase, and phenylacetyl coenzyme A ligase (Ho et al., 2018). It is widely postulated that these enzymes participate in a cascade of processes encompassing the depolymerization of the polymer into styrene, the oxidation of styrene into phenylacetate, and the assimilation of phenylacetate into the Krebs cycle.

3.7.5 Nylon-degrading enzymes

Several enzymes capable of degrading nylon have been found in both fungal and bacterial sources (Nomura et al., 2001; Yamano et al., 2019). One example of an enzyme with the ability to modify Nylon 6 is the MnP derived from a white-rot fungus known as strain IZU-154. In a study conducted by Deguchi et al. (1998), it was demonstrated that this enzyme is capable of removing the surface layer of Nylon 6 and inducing the creation of distinct horizontal grooves within the polymer. Peroxidases encompass a class of enzymes that possess a heme prosthetic group, facilitating their ability to facilitate the oxidation of a diverse range of organic and inorganic substrates. These enzymatic reactions rely on the utilization of hydrogen peroxide as the electron acceptor. Furthermore, it has been demonstrated that the metabolic process of 6-aminohexanoate, which serves as an intermediary compound in the production of nylon, is facilitated by three distinct hydrolases found in *Flavobacterium* and *Pseudomonas strains* (Negoro, 2000). The enzymes, namely, 6-aminohexanoate-cyclic-dimer hydrolase, 6-aminohexanoate-dimer hydrolase, and endo-type 6-aminohexanoate-oligomer hydrolase, were found to work together sequentially to transform the nylon intermediate into its monomer, 6-

aminohexanoate. This conversion process occurred under conditions of moderate temperature and a pH range from neutral to alkaline. Additional research has subsequently revealed the existence of enzymes exhibiting characteristics comparable with those previously stated in *Arthrobacter* sp. KI72 (Yasuhira et al., 2007).

3.7.6 Polylactic acid–degrading enzymes

In a separate investigation, the impact of four distinct enzymes (proteinase K, protease, esterase, and lipase) on the degradation process of flax fiber reinforced PLA was examined. The application of SEM in morphological investigations of flax fiber reinforced PLA revealed that the enzyme proteinase K exhibited the most significant influence on the degradability of the polymers. A significant increase in mass loss, approximately 52%, was seen in the case of PLA reinforced with 30% flax fiber, in comparison with a mass loss of just 21% in the case of pure PLA polymer. The use of flax fiber resulted in an enhancement of the polymer's biodegradability. Proteinase K induced the most significant alteration in the overall structure of the PLA polymer. According to Stepczyńska and Rytlewski (2018), the breakdown of polymers was minimally affected by the presence of protease and lipase enzymes.

The lipase enzyme derived from Candida rugosa and the proteinase K enzyme derived from *Tritirachium* album were shown to induce degradation of PLA polymer and its nanocomposites, which were reinforced with cellulose nanocrystals. According to the findings of the investigation, it was observed that the enzyme lipase exhibited limited efficacy in facilitating the disintegration of the polymer. Conversely, the outcomes obtained using the enzyme proteinase K had encouraging potential. The generation of lactic acid during the course of the reaction resulted in a reduction in the pH of the reaction mixture, leading to the deterioration of the enzymes. The degrading process of the polymer was shown to be more favorable under conditions of reduced ionic strength.

Previous studies have documented the ability of lipase and PU esterase enzymes derived from *Rhizopus delemar* and *Comamonas acidovorans* to degrade low–-molecular-weight PLA. Additionally, *Amycolatopsis* sp. has been found to digest high–molecular-weight PLA (Masaki et al., 2005).

The utilization of cellulose nanocrystals in conjunction with PLA polymer facilitated the enhancement of degradation kinetics, as evidenced by the observed degradation of the polymer sample within a span of 3 days (Hegyesi et al., 2019). The enzyme proteinase K was employed for the breakdown of blend films composed of PBS/PLA and reinforced with benzyl peroxide. The films comprising a blend exhibited a visually smooth surface prior to conducting degradation investigations, as observed by the utilization of SEM. According to Hu et al. (2018), the film's surface exhibited the emergence of microscopic fractures and saw an increase in roughness following a 12-hour treatment with the enzyme.

Furthermore, the roughness continued to escalate after undergoing degradation for a period of 24 hours.

3.7.7 Other plastic-degrading enzymes studies

The degradation of polyhydroxybutyrate (PHB), PBS, and polyethylene succinate (PES) has been the subject of investigation, with a particular focus on the use of a PHB depolymerase as described by Jung et al. (2018). The enzyme was isolated from *Aspergillus fumigatus* and subsequently subjected to cloning and purification procedures (Kasuya et al., 1998). The enzyme in question has been documented as an extracellular enzyme that exhibits activity towards PHB, PBS, and PES. Its optimal activity is observed at a pH of 6.5 at a temperature of 55°C. The enzyme isolation procedure involved the selection of bacteria that exhibited a distinct zone on agar plates with PHB, PBS, and PES as the sole carbon source, as described by Hoang et al. (2007). According to Tokiwa and Calabia (2004), the presence of a distinct clear zone demonstrated the strain's capability to degrade a range of plastic materials. Additionally, this strain was employed for the isolation of the depolymerase enzyme. A significant reduction in weight, around 95%, was documented for the plastic films after being subjected to a 17-hour incubation period with depolymerase, as reported by Ohtaki et al. (2006). According to Dickinson (1998), the films exhibited a transition in surface topology, shifting from a smooth look to a fibrous appearance characterized by the presence of potholes measuring 3–5 mm. This change in surface morphology suggests a significant level of film deterioration.

The enzymatic degradation of PBTF (poly(1,4-butylene 2,5-thiophenedicarboxylate)) and PBF (poly(1,4-butylene 2,5-furandicarboxylate)) films was conducted using cutinase enzymes derived from HiC and the thermostable *Thermobifida cellulosilytica* cutinase 1 (Cut). The enzymatic hydrolysis process was conducted for a duration of approximately 72 hours, utilizing an enzyme concentration of 5 μM in a buffer solution consisting of 1 M K_2HPO_4/KH_2PO_4. According to previous studies, it has been shown that maintaining a pH level of 8 yields optimal outcomes for film deterioration. The videos provided proof of a weight loss of approximately 20%, therefore demonstrating the effectiveness of the enzyme activity. The degradation rate of PBF was observed to be somewhat higher compared with that of PBTF. In a study conducted by Giglia et al. (2019), it was observed that the enzymatic activity of both HiC and Cut was enhanced at elevated temperatures, namely, at 65°C as opposed to 50°C. Furthermore, HiC exhibited a more rapid kinetic rate of hydrolysis when compared with Cut.

Knott et al. (2020) reported a notably synergistic interaction between the PETase enzyme and the MHETase enzyme derived from *I. sakaiensis* in converting amorphous PET film into its constituent monomeric units, namely TPA and ethylene glycol. The degree of hydrolysis of PET at a temperature of 30°C was examined for 96 hours in the presence of two enzymes at varying doses. The

MHETase enzyme exhibited no discernible activity in the breakdown of PET film. When MHETase is introduced into the process alongside the PETase enzyme, the release of the product (TPA and ethylene glycol) is found to be directly proportional to the amount of PETase present. However, the level of product release is significantly higher when MHETase is present in addition to PETase, compared with when PETase is used alone. The release of constituent monomers was found to rise as the concentration of both enzymes rose, suggesting that these processes were restricted by enzyme availability rather than substrate availability. The findings of the synergy study indicate that the breakdown of PETase enzyme is influenced by the presence of MHETase, even when MHETase is present in low quantities compared with PETase.

The utilization of PU as a substrate and carbon source, as well as its degradation, has been demonstrated by *Pestalotiopsis microspora*, which possesses serine hydrolase (Russell et al., 2011).

3.8 Conclusion and future aspects

Microbial enzymes play a crucial role as significant environmental agents in facilitating the process of biodegradation. The phenomenon of biodegradation entails the transformation of carbon present in polymer chains into either smaller biomolecules or into carbon dioxide and water (Mir et al., 2017). Consequently, the introduction of organic matter into the soil enhances its fertility while simultaneously mitigating the accumulation of plastics, thereby resulting in a reduction in waste management expenses. Microbial enzymes are of significant importance in the treatment of plastic waste. The implementation of novel techniques and new solutions plays a crucial role in mitigating the production of nonbiodegradable and nonreusable plastics. Additionally, enhancing target enzymes or microbiomes involved in plastic biodegradation might serve as effective instruments for managing plastic waste (Gricajeva et al., 2022).

Enzymes exhibit significant potential for the degradation of both biodegradable and nonbiodegradable plastic polymers, which contribute to environmental pollution and represent a risk to all terrestrial life forms. Numerous scientists continue to investigate novel and enhanced strategies for the degradation of diverse kinds of plastics through the utilization of enzymes derived from various microbial sources. The degradation of plastic polymers has been mostly attributed to hydrolytic enzymes, as indicated by the majority of studies. Recent studies in this field have demonstrated that the current focus should be on augmenting the efficacy of enzymes known to facilitate the degradation of different plastic materials, in order to establish enzymatic plastic degradation as a viable commercial procedure.

One approach to improve the stability of plastic-degrading enzymes in challenging environmental circumstances, such as high temperatures or extreme cold, involves manipulating the active site of these enzymes. Site-directed

mutagenesis is a technique that can be utilized to modify the structure and functional groups of an enzyme's active site. This alteration enables the enzyme to readily accommodate flexible polymers and exhibit functionality across a broad variety of temperature and pH conditions. Furthermore, the utilization of a composite bifunctional system consisting of two enzymes exhibits the capacity to tolerate multiple varieties of plastics, hence expanding the scope of plastic degradability inside a singular experimental setting. The discovery occurred when a bifunctional lipase-cutinase enzyme system was utilized for the degradation of PCL polymeric material.

The standardization and optimization of the isolation procedure for microbial species with plastic-degrading enzymes is of paramount importance. This is necessary to facilitate the efficient screening of pure breeding colonies of these microorganisms within a minimal time frame. The implementation of a standardized initial phase has the potential to enhance the generation of enzymes from pure colonies, hence expediting the degradation process of plastic polymers.

References

Abd El-Rehim, H.A., Hegazy, E.-S.A., Ali, A.M., Rabie, A.M., 2004. Synergistic effect of combining UV-sunlight–soil burial treatment on the biodegradation rate of LDPE/starch blends. Journal of Photochemistry and Photobiology A: Chemistry 163 (3), 547–556. Available at https://doi.org/10.1016/j.jphotochem.2004.02.003.

Alfieri, B., Alfieri, M., Kelly, M., Kilcoyne, S., Poprik, L., Sanyal, A., et al., 2022. The role of Ideonella sakaiensis PETase in the degradation of PET plastics: a structural comparison of the wild type and S238F/W159H double mutant. The FASEB Journal 36 (S1). Available at https://doi.org/10.1096/fasebj.2022.36.s1.l8070.

Ali, S.S., Elsamahy, T., Al-Tohamy, R., Zhu, D., Mahmoud, Y.A.-G., Koutra, E., et al., 2021. Plastic wastes biodegradation: mechanisms, challenges and future prospects. Science of The Total Environment 780, 146590. Available at https://doi.org/10.1016/j.scitotenv.2021.146590.

Alisch-Mark, M., Herrmann, A., Zimmermann, W., 2006. Increase of the hydrophilicity of polyethylene terephthalate fibers by hydrolases from Thermomonospora fusca and Fusarium solani f. sp. pisi. Biotechnology Letters 28, 681–685.

Allen, A.B., Hilliard, N.P., Howard, G.T., 1999. Purification and characterization of a soluble polyurethane degrading enzyme from *Comamonas acidovorans*. Int Biodeterior Biodegrad 43, 37–41.

Amin, M., Bhatti, H.N., Bilal, M., 2022. Kinetic and thermodynamic characterization of lipase from *Aspergillus melleus* and its biocatalytic performance for degradation of poly (ε-caprolactone). Journal of Chemical Technology & Biotechnology 97, 446–454.

Amobonye, A., Bhagwat, P., Singh, S., Pillai, S., 2021. Plastic biodegradation: frontline microbes and their enzymes. Science of The Total Environment 759, 143536. Available at https://doi.org/10.1016/j.scitotenv.2020.143536.

Anbalagan, S., Venkatakrishnan, H.R., Ravindran, J., Sathyamoorthy, J., Rangabashyam, K.A., Ragini, Y.P., et al., 2021. Hydrolytic degradation of polyethylene terephthalate by

cutinase enzyme derived from fungal biomass-molecular characterization. Biointerface Research in Applied Chemistry 12, 653–667.

Aris, M.H., Annuar, M.S.M., Ling, T.C., 2016. Lipase-mediated degradation of poly-ε-caprolactone in toluene: behavior and its action mechanism. Polymer Degradation and Stability 133, 182–191.

Artham, T., Doble, M., 2008. Biodegradation of aliphatic and aromatic polycarbonates. Macromolecular Bioscience 8 (1), 14–24. Available at https://doi.org/10.1002/mabi.200700106.

Austin, H.P., Allen, M.D., Donohoe, B.S., Rorrer, N.A., Kearns, F.L., Silveira, R.L., et al., 2018. Characterization and engineering of a plastic-degrading aromatic polyesterase. Proceedings of the National Academy of Sciences of the United States of America 115 (19), E4350. Available at https://doi.org/10.1073/pnas.1718804115, http://www.pnas.org/content/pnas/115/19/E4350.full.pdf.

Barragán, D.H., Pelacho, A.M., Closas, L.M., 2016. Degradation of agricultural biodegradable plastics in the soil under laboratory conditions. Soil Research 54, 216–224.

Barth, M., Oeser, T., Wei, R., Then, J., Schmidt, J., Zimmermann, W., 2015. Effect of hydrolysis products on the enzymatic degradation of polyethylene terephthalate nanoparticles by a polyester hydrolase from Thermobifida fusca. Biochemical Engineering Journal 93, 222–228. Available at https://doi.org/10.1016/j.bej.2014.10.012.

Barth, M., Honak, A., Oeser, T., Wei, R., Belisário-Ferrari, M.R., Then, J., et al., 2016. A dual enzyme system composed of a polyester hydrolase and a carboxylesterase enhances the biocatalytic degradation of polyethylene terephthalate films. Biotechnology Journal 11 (8), 1082–1087. Available at https://doi.org/10.1002/biot.201600008, http://onlinelibrary.wiley.com/journal/10.1002/(ISSN)1860-7314.

Bhardwaj, H., Gupta, R., Tiwari, A., 2013. Communities of microbial enzymes associated with biodegradation of plastics. Journal of Polymers and the Environment 21, 575–579.

Bholay, A.D., Borkhataria, V., Jadhav, U., Palekar, S., Dhalkari, V., Nalawade, P.M., et al., 2012. Bacterial lignin peroxidase: a tool for bioleccching and biodegra- dation of industrial effluents. Universal Journal of Environmental Research and Technology 2, 58–64.

Billig, S., Oeser, T., Birkemeyer, C., Zimmermann, W., 2010. Hydrolysis of cyclic poly (ethylene terephthalate) trimers by a carboxylesterase from Thermobifida fusca KW3. Applied Microbiology and Biotechnology 87.

Bo, R.K., Soo, B.K., Hyun, A.S., Tae, K.L., 2019. Accelerating the biodegradation of high-density polyethylene (HDPE) using Bjerkandera adusta TBB-03 and lignocellulose substrates. MDPI Microorganisms 7, 304.

Booth, A.M., Kubowicz, S., Beegle-Krause, C.J., Skancke, J., Nordam, T., Landsem, E., et al., 2017. Microplastic in global and norwegian marine environments: distributions, degradation mechanisms and transport. Miljødirektoratet 918, 1–147.

Brunner, I., Fischer, M., Rüthi, J., Stierli, B., Frey, B., Aroca, R., 2018. Ability of fungi isolated from plastic debris floating in the shoreline of a lake to degrade plastics. PLoS One 13 (8), e0202047. Available at https://doi.org/10.1371/journal.pone.0202047.

Carniel, A., Valoni, E., Nicomedes, J., Conceição Gomes, A., Castro, A.M., 2016. Lipase from Candida antarctica (CALB) and cutinase from *Humicola insolens* act synergistically for PET hydrolysis to terephthalic acid. Process Biochemistry 59, 84–90.

Carniel, A., Valoni, É., Nicomedes, J., Gomes, A.D.C., Castro, A.M.D., 2017. Lipase from Candida antarctica (CALB) and cutinase from Humicola insolens act synergistically for

PET hydrolysis to terephthalic acid. Process Biochemistry 59, 84–90. Available at https://doi.org/10.1016/j.procbio.2016.07.023.

Carrott, P.J., Carrott, M.R., 2007. Lignin-from natural adsorbent to activated carbon: a review. Bioresource Technology 98.

Chen, S., Su, L., Chen, J., Wu, J., 2013. Cutinase: characteristics, preparation, and application. Biotechnology Advances 31 (8), 1754–1767. Available at https://doi.org/10.1016/j.biotechadv.2013.09.005.

Chinaglia, S., Tosin, M., Degli-Innocenti, F., 2018. Biodegradation rate of biodegradable plastics at molecular level. Polym.Polymer Degradation and Stability 147, 237–244.

Closas, L.M., Costa, J., Cirujeda, A., Aibar, J., Zaragoza, C., Pardo, A., et al., 2016. Above-soil and in-soil degradation of oxo- and bio-degradable mulches: a qualitative approach. Soil Research 54, 225–236.

Cosgrove, L., McGeechan, P.L., Robson, G.D., Handley, P.S., 2007. Fungal communities associated with degradation of polyester polyurethane in soil. Applied and Environmental Microbiology 73 (18), 5817–5824. Available at https://doi.org/10.1128/AEM.01083-07.

Danso, D., Chow, J., Streit, W.R., Drake, H.L., 2019. Plastics: environmental and biotechnological perspectives on microbial degradation. Applied and Environmental Microbiology 85 (19). Available at https://doi.org/10.1128/aem.01095-19.

Dashtban, M., Schraft, H., Syed, T.A., Qin, W., 2010. Fungal biodegradation and enzymatic modification of lignin. International Journal of Biochemistry and Molecular Biology 1 (1), 36–50. Available at http://www.ijbmb.org/files/IJBMB1004005.pdf.

de Dicastillo, C.L., Velásquez, E., Rojas, A., Guarda, A., Galotto, M.J., 2020. The use of nanoadditives within recycled polymers for food packaging: properties, recyclability, and safety. Comprehensive Reviews in Food Science and Food Safety 19, 1760–1776.

Deguchi, T., Kitaoka, Y., Kakezawa, M., Nishida, T., 1998. Purification and characterization of a nylon-degrading enzyme. Applied and Environmental Microbiology 64 (4), 1366–1371. Available at https://doi.org/10.1128/aem.64.4.1366-1371.1998.

Dickinson, E., 1998. Proteins at interfaces and in emulsions stability, rheology and interactions. Journal of the Chemical Society, Faraday Transactions 94 (12), 1657–1669. Available at https://doi.org/10.1039/a801167b.

Eberl, A., Heumann, S., Brückner, T., Araujo, R., Cavaco-Paulo, A., Kaufmann, F., et al., 2009. Enzymatic surface hydrolysis of poly(ethylene terephthalate) and bis (benzoyloxyethyl) terephthalate by lipase and cutinase in the presence of surface active molecules. Journal of Biotechnology 143 (3), 207–212. Available at https://doi.org/10.1016/j.jbiotec.2009.07.008.

Fotopoulou, K.N., Karapanagioti, H.K., 2017. Degradation of various plastics in the environment. In: Hazardous Chemicals Associated With Plastics in the Marine Environment. The Handbook of Environmental Chemistry, vol. 78. Springer Science and Business Media LLC, pp. 71–92. Available at https://doi.org/10.1007/698_2017_11

Furukawa, M., Kawakami, N., Tomizawa, A., Miyamoto, K., 2019. Efficient degradation of poly(ethylene terephthalate) with Thermobifida fusca cutinase exhibiting improved catalytic activity generated using mutagenesis and additive-based approaches. Scientific Reports 9 (1). Available at https://doi.org/10.1038/s41598-019-52379-z.

Gan, Z., Zhang, H., 2019. PMBD: a comprehensive plastics microbial biodegradation database. Database 2019. Available at https://doi.org/10.1093/database/baz119.

Gan, Z., Abe, H., Doi, Y., 2001. Crystallization, melting, and enzymatic degradation of biodegradable poly (butylene succinate-co-14 mol % ethylene succinate) copolyester. Biomacromolecules 2, 313–321.

Ganesh, P., Dineshraj, D., Yoganathan, K., 2017. Production and screening of depolymerising enzymes by potential bacteria and fungi isolated from plastic waste dump yard sites. International Journal of Applied Research 3 (3), 693–695.

Gaytán, I., Sánchez-Reyes, A., Burelo, M., Vargas-Suárez, M., Liachko, I., Press, M., et al., 2020. Degradation of recalcitrant polyurethane and xenobiotic additives by a selected landfill microbial community and its biodegradative potential revealed by proximity ligation-based metagenomic analysis. Frontiers in Microbiology 10. Available at https://doi.org/10.3389/fmicb.2019.02986, https://www.frontiersin.org/journals/microbiology#.

Giglia, M., Quartinellob, F., Soccioc, M., Pellisb, A., Lottic, N., Georg, M.G., et al., 2019. Enzymatic hydrolysis of poly(1,4-butylene 2,5-thiophenedicarboxylate) (PBTF) and poly (1,4-butylene 2,5-furandicarboxylate) (PBF) films: a comparison of mechanisms. Environment International 130.

Glaser, J.A., 2019. Biological degradation of polymers in the environment. Plastics in the Environment. IntechOpen.

Gomez-Mendez, L.D., Moreno-Bayona, D.A., Poutou-Pinales, R.A., Salcedo-Reyes, J.C., Pedroza- Rodriguez, A.M., Vargas, A., et al., 2018. Biodeterioration of plasma pretreated LDPE sheets by Pleurotus ostreatus. PLoS One 13 (9), e0203786. Available at https://doi.org/10.1371/journal.pone.0203786.

Gricajeva, A., Nadda, A.K., Gudiukaite, R., 2022. Insights into polyester plastic biodegradation by carboxyl ester hydrolases. Journal of Chemical Technology and Biotechnology (Oxford, Oxfordshire: 1986) 97, 359–380.

Gross, R.A., Kalra, B., 2002. Biodegradable polymers for the environment. Science (New York, N.Y.) 297 (5582), 803–807. Available at https://doi.org/10.1126/science.297.5582.803.

Gu, J.D., 2003. Microbiological deterioration and degradation of synthetic polymeric materials: recent research advances. International Biodeterioration and Biodegradation 52 (2), 69–91. Available at https://doi.org/10.1016/S0964-8305(02)00177-4, www.elsevier.com/inca/publications/store/4/0/5/8/9/9.

Hegyesi, N., Zhang, Y., Kohári, A., Polyák, P., Sui, X., Pukánszky, B., 2019. Enzymatic degradation of PLA/cellulose nanocrystal composites. Industrial Crops and Products 141. Available at https://doi.org/10.1016/j.indcrop.2019.111799, www.elsevier.com/inca/publications/store/5/2/2/8/2/5.

Heredia, A., 2003. Biochemical and biophysical characteristics of cutin, a plant barrier biopolymer. Biochimica et Biophysica Acta 1620, 1–7. Available at https://doi.org/10.1016/S0304-4165(02)00510-X.

Ho, B.T., Roberts, T.K., Lucas,, S., 2018. An overview on biodegradation of polystyrene and modified polystyrene: the microbial approach. Critical Reviews in Biotechnology 38, 308–320. Available at https://doi.org/10.1080/07388551.2017.1355293.

Hoang, K.C.,, Tseng, M., Shu, W.J., 2007. Degradation of polyethylene succinate (PES) by a new thermophilic Microbispora strain. Biodegradation 18 (3), 333–342.

Hofrichter, M., Ullrich, R., 2006. Heme-thiolate haloperoxidases: versatile biocatalysts with biotechnological and environmental significance. Applied Microbiology and Biotechnology 71 (3), 276–288. Available at https://doi.org/10.1007/s00253-006-0417-3.

Howard, G.T., Ruiz, C., Hilliard, N.P., 1999. Growth of *Pseudomonas chlororaphis* on apolyester-polyurethane and the purification andcharacterization of a polyurethanase-esterase enzyme. International Biodeterioration & Biodegradation 43, 7–12.

Howard, G.T., Norton, W.N., Burks, T., 2012. Growth of *Acinetobacter gerneri P7* on polyurethane and the purification and characterization of a polyurethanase enzyme. Biodegradation 23, 561–573.

Hu, X., Gao, Z., Wang, Z., Su, T., Yang, L., Li, P., 2016. Enzymatic degradation of poly (butylene succinate) by cutinase cloned from Fusarium solani. Polymer Degradation and Stability 134, 211–219. Available at https://doi.org/10.1016/j.polymdegradstab.2016.10.012.

Hu, X., Su, T., Li, P., Wang, Z., 2018. Blending modification of PBS/PLA and its enzymatic degradation. Polymer Bulletin 75 (2), 533–546. Available at https://doi.org/10.1007/s00289-017-2054-7.

Iiyoshi, Y., Tsutsumi, Y., Nishida, T., 1998. Polyethylene degradation by lignin-degrading fungi and manganese peroxidase. Journal of Wood Science 44, 222–229.

Ishigaki, T., Sugano, W., Ike, M., Fujita, M., 2000. Enzymatic degradation of cellulose acetate plastic by novel degrading bacterium Bacillus sp. S2055. Journal of Bioscience and Bioengineering 90 (4), 400–405. Available at https://doi.org/10.1016/s1389-1723(01)80008-6.

Jabloune, R., Khalil, M., Ben Moussa, I.E., Simao-Beaunoir, A.M., Lerat, S., Brzezinski, R., et al., 2020. Enzymatic degradation of p-nitrophenyl esters, polyethylene terephthalate, cutin, and suberin by Sub1, a suberinase encoded by the plant pathogen Streptomyces scabies. Microbes and Environments 35 (1). Available at https://doi.org/10.1264/jsme2.ME19086.

Jeon, H.J., Kim, M.N., 2015. Functional analysis of alkane hydroxylase system derived from Pseudomonas aeruginosa E7 for low molecular weight polyethylene biodegradation. International Biodeterioration and Biodegradation 103, 141–146. Available at https://doi.org/10.1016/j.ibiod.2015.04.024, www.elsevier.com/inca/publications/store/4/0/5/8/9/9.

Jia, H., Zhang, M., Weng, Y., Zhao, Y., Li, C., Kanwal, A., 2021. Degradation of poly (butylene adipate-co-terephthalate) by Stenotrophomonas sp. YCJ1 isolated from farmland soil. Journal of Environmental Sciences 103, 50–58. Available at https://doi.org/10.1016/j.jes.2020.10.001.

Joo, S., Cho, I.J., Seo, H., Son, H.F., Sagong, H.-Y., Joo Shin, T., et al., 2018. Structural insight into molecular mechanism of poly(ethylene terephthalate) degradation. Nature Communications 9 (1). Available at https://doi.org/10.1038/s41467-018-02881-1.

Jung, H.W., Yang, M.K., Su, R.-C., 2018. Purification, characterization, and gene cloning of an Aspergillus fumigates polyhydroxy butyrate depolymerase used for degradation of polyhydroxybutyrate, polyethylene succinate, and polybutylene succinate. Polymer Degradation and Stability 154, 186–194.

Kamboj, M., 2016. Degradation of plastics for clean environment. International Journal of Advanced Research in Engineering and Applied Sciences 5 (3).

Kasuya, K., Takagi, K., Ishiwatari, S., Yoshida, Y., Doi, Y., 1998. Biodegradabilities of various aliphatic polyesters in natural waters. Polymer Degradation and Stability 59 (1–3), 327–332. Available at https://doi.org/10.1016/s0141-3910(97)00155-9.

Khan, I., Dutta, J.R., Ganesan, R., 2017. *Lactobacillus* sps. lipase mediated poly (epsiloncaprolactone) degradation. International Journal of Biological Macromolecules 95, 126–131.

Kijchavengkul, T., Auras, R., Rubino, M., Selke, S., Ngouajio, M., Fernandez, R.T., 2010. Biodegradation and hydrolysis rate of aliphatic aromatic polyester. Polymer Degradation and Stability 95 (12), 2641–2647. Available at https://doi.org/10.1016/j.polymdegradstab.2010.07.018.

Knott, B.C., Erickson, E., Allen, M.D., Gado, J.E., Graham, R., Kearns, F.L., et al., 2020. Characterization and engineering of a two-enzyme system for plastics depolymerization. Proceedings of the National Academy of Sciences of the United States of America 117 (41), 25476–25485. Available at https://doi.org/10.1073/pnas.2006753117, https://www.pnas.org/content/pnas/117/41/25476.full.pdf.

Kolattukudy, P.E., Brown, L., 1975. Fate of naturally occurring epoxy acids: a soluble epoxide hydrase, which catalyzes cis hydration, from Fusarium solani pisi. Archives of Biochemistry and Biophysics 166 (2), 599–607. Available at https://doi.org/10.1016/0003-9861(75)90425-7.

Kolattukudy, P.E., Purdy, R.E., Maiti, I.B., 1981. Cutinases from fungi and pollen. In: Lowenstein, J.M. (Ed.), Methods in Enzymology. Academic Press, New York, pp. 652.

Kour, H., Khan, S.S., Kour, D., Rasool, S., Sharma, Y.P., Rai, P.K., et al., 2023. Microbes mediated plastic degradation: a sustainable approach for environmental sustainability. Journal of Applied Biology and Biotechnology 11 (3), 9–19. Available at https://doi.org/10.7324/JABB.2023.110515, jabonline.in/index.php.

Krueger, M.C., Harms, H., Schlosser, D., 2015. Prospects for microbiological solutions to environmental pollution with plastics. Applied Microbiology and Biotechnology 99, 8857–8874.

Kyrikou, I., Briassoulis, D., 2007. Biodegradation of agricultural plastic films: a critical review. Journal of Polymers and the Environment 15, 125–150.

Liebminger, S., Eberl, A., Sousa, F., Heumann, S., Fischer-Colbrie, G., Cavaco-Paulo, A., et al., 2007. Hydrolysis of PET and bis-(benzoyloxyethyl) terephthalate with a new polyesterase from Penicillium citrinum. Biocatalysis and Biotransformation 25 (2–4), 171–177. Available at https://doi.org/10.1080/10242420701379734.

Li, F., Hu, X., Guo, Z., Wang, Z., Wang, Y., Liu, D., et al., 2011. Purification and characterization of a novel poly(butylene succinate)-degrading enzyme from Aspergillus sp. XH0501-a. World Journal of Microbiology and Biotechnology 27 (11), 2591–2596. Available at https://doi.org/10.1007/s11274-011-0731-5.

Liu, M., Zhang, T., Long, L., Zhang, R., Ding, S., 2019. Efficient enzymatic degradation of poly (ε-caprolactone) by an engineered bifunctional lipase-cutinase. Polymer Degradation and Stability 160, 120–125. Available at https://doi.org/10.1016/j.polymdegradstab.2018.12.020.

Loredo-Treviño, A., García, G., Velasco-Téllez, A., Rodríguez-Herrera, R., Aguilar, C.N., 2011. Polyurethane as substrate for fungal strains. Advances in Bioscience and Biotechnology 02 (02), 52–58. Available at https://doi.org/10.4236/abb.2011.22009.

Lülsdorf, N., Vojcic, L., Hellmuth, H., Weber, T.T., Mußmann, N., Martinez, R., et al., 2015. A first continuous 4-aminoantipyrine (4-AAP)-based screening system for directed esterase evolution. Applied Microbiology and Biotechnology 99.

Maeda, H., Yamagata, Y., Abe, K., Hasegawa, F., Machida, M., Ishioka, R., et al., 2005. Purification and characterization of a biodegradable plastic-degrading enzyme from Aspergillus oryzae. Applied Microbiology and Biotechnology 67 (6), 778–788. Available at https://doi.org/10.1007/s00253-004-1853-6.

Magnin, A., Pollet, E., Perrin, R., Ullmann, C., Persillon, C., Phalip, V., et al., 2019. Enzymatic recycling of thermoplastic polyurethanes: synergistic effect of an esterase and an amidase and recovery of building blocks. Waste Management 85, 141–150. Available at https://doi.org/10.1016/j.wasman.2018.12.024.

Magnin, A., Pollet, E., Phalip, V., Avérous, L., 2020. Evaluation of biological degradation of polyurethanes. Biotechnology Advances 39, 107457. Available at https://doi.org/10.1016/j.biotechadv.2019.107457.

Marija, S.N.,, Jasna, D., 2001. Synthesis and characterization of biodegradable poly (butylene succinate-co-butylene adipate). Polymer Degradation and Stability 74, 263–270.

Masaki, K., Kamini, N.R., Ikeda, H., Iefuji, H., 2005. Cutinase-like enzyme from the yeast Cryptococcus sp. strain S-2 hydrolyzes polylactic acid and other biodegradable plastics. Applied and Environmental Microbiology 71 (11), 7548–7550. Available at https://doi.org/10.1128/AEM.71.11.7548-7550.2005.

Mir, S., Asghar, B., Khan, A.K., Rashid, R., Shaikh, A.J., Khan, R.A., et al., 2017. The effects of nanoclay on thermal, mechanical and rheological properties of LLDPE/chitosan blend. Journal of Polymer Engineering 37 (2), 143–149. Available at https://doi.org/10.1515/polyeng-2015-0350, http://www.reference-global.com/loi/polyeng.

Mohan, A.J., Sekhar, V.C., Bhaskar, T., Nampoothiri, K.M., 2016. Microbial assisted High Impact Polystyrene (HIPS) degradation. Bioresource Technology 213, 204–207. Available at https://doi.org/10.1016/j.biortech.2016.03.021, www.elsevier.com/locate/biortech.

Mohanan, N., Wong, C.H., Budisa, N., Levin, D.B., 2022. Characterization of polymer degrading lipases, LIP1 and LIP2 from Pseudomonas chlororaphis PA23. Frontiers in Bioengineering and Biotechnology 10. Available at https://doi.org/10.3389/fbioe.2022.854298, http://journal.frontiersin.org/journal/bioengineering-and-biotechnology#archive.

Mukai, K., Doi, Y., Sema, Y., Tomita, K., 1993. Substrate specificities in hydrolysis of polyhydroxyalkanoates by microbial esterases. Biotechnology Letters 15 (6), 601–604. Available at https://doi.org/10.1007/bf00138548.

Müller, R.J., Schrader, H., Profe, J., Dresler, K., Deckwer, W.D., 2005. Enzymatic degradation of poly(ethylene terephthalate): rapid hydrolyse using a hydrolase from T. fusca. Macromolecular Rapid Communications 26 (17), 1400–1405. Available at https://doi.org/10.1002/marc.200500410.

Munari, 2019. Enzymatic hydrolysis of poly (1,4-butylene 2,5-thiophenedi-carboxylate) (PBTF) and poly(1,4-butylene 2,5-furandicarboxylate) (PBF) films: a comparison of mechanisms. Environment International 130.

Nag, M., Lahiri, D., Dutta, B., Jadav, G., Ray, R.R., 2021. Biodegradation of used polyethylene bags by a new marine strain of Alcaligenes faecalis LNDR-1. Environmental Science and Pollution Research 28 (30), 41365–41379. Available at https://doi.org/10.1007/s11356-021-13704-0, https://link.springer.com/journal/11356.

Nair, S., Kumar, P., 2007. Molecular characterization of a lipase- producing *Bacillus pumilus* strain (NMSN-1d) utilizing colloidal water-dispersible polyurethane. World Journal of Microbiology and Biotechnology 23, 1441–1449.

Nakajima-Kambe, T., Edwinoliver, N.G., Maeda, H., Thirunavukarasu, K., Gowthaman, M.K., Masaki, K., et al., 2012. Purification, cloning and expression of an Aspergillus niger lipase for degradation of poly(lactic acid) and poly(ε-caprolactone). Polymer Degradation and Stability 97 (2), 139–144. Available at https://doi.org/10.1016/j.polymdegradstab.2011.11.009.

Nakkabi, A., Sadiki, M., Fahim, M., Ittobane, N., Ibnsouda Koraichi, S., Barkai, H., et al., 2015. Biodegradation of poly(ester urethane)s by Bacillus subtilis. International Journal of Environmental Research 9 (1), 157–162. Available at http://ijer.ut.ac.ir/pdf_885_e90b943efc9d870f515b6d10d3e98dd5.html.

Negoro, S., 2000. Biodegradation of nylon oligomers. Applied Microbiology and Biotechnology 54 (4), 461–466. Available at https://doi.org/10.1007/s002530000434.

Nikolaivits, E., Taxeidis, G., Gkountela, C., Vouyiouka, S., Maslak, V., Nikodinovic-Runic, J., et al., 2022. A polyesterase from the Antarctic bacterium Moraxella sp. degrades highly crystalline synthetic polymers. Journal of Hazardous Materials 434, 128900. Available at https://doi.org/10.1016/j.jhazmat.2022.128900.

Nomura, N., Deguchi, T., Shigeno-Akutsu, Y., Nakajima-Kambe, T., Nakahara, T., 2001. Gene structures and catalytic mechanisms of microbial enzymes able to blodegrade the synthetic solid polymers nylon and polyester polyurethaoe. Biotechnology and Genetic Engineering Reviews 18 (1), 125–147. Available at https://doi.org/10.1080/02648725.2001.10648011.

Nunes, C.S., Kunamneni, A., 2018. Laccases-properties and applications. Enzymes in Human and Animal Nutrition: Principles and Perspectives. Elsevier Inc., France, pp. 133–161. Available at https://www.sciencedirect.com/book/9780128054192/enzymes-in-human-and-animal-nutrition 10.1016/B978-0-12-805419-2.00007-1.

Oda, Y., Yonetsu, A., Urakami, T., Tonomura, K., 2000. Degradation of polylactide by commercial proteases. Journal of Polymers and the Environment 8 (1), 29–32. Available at https://doi.org/10.1023/A:1010120128048.

Oeser, T., Wei, R., Baumgarten, T., Billig, S., Föllner, C., Zimmermann, W., 2010. High level expression of a hydrophobic poly(ethylene terephthalate)-hydrolyzing carboxylesterase from Thermobifida fusca KW3 in Escherichia coli BL21(DE3). Journal of Biotechnology 146 (3), 100–104. Available at https://doi.org/10.1016/j.jbiotec.2010.02.006.

Ohtaki, S., Maeda, H., Takahashi, T., Yamagata, Y., Hasegawa, F., Gomi, K., et al., 2006. Novel hydrophobic surface binding protein, HsbA, produced by Aspergillus oryzae. Applied and Environmental Microbiology 72 (4), 2407–2413. Available at https://doi.org/10.1128/aem.72.4.2407-2413.2006.

Osma, J.F., Toca-Herrera, J.L., Rodríguez-Couto, S., 2010. Uses of laccases in the food industry. Enzyme Research 2010. Available at https://doi.org/10.4061/2010/918761 918761.

Ozsagiroglu, E., Iyisan, B., Guvenilir, Y.A., 2012. Biodegradation and characterization studies of different kinds of polyurethenes with several enzyme solutions. Polish Journal of Environmental Studies 6, 1777–1782.

O'Neill, A., Araújo, R., Casal, M., Guebitz, G., Cavaco-Paulo, A., 2007. Effect of the agitation on the adsorption and hydrolytic efficiency of cutinases on polyethylene terephthalate fibres. Enzyme and Microbial Technology 40 (7), 1801–1805. Available at https://doi.org/10.1016/j.enzmictec.2007.02.012.

Paci, M., La Mantia, F.P., 1998. Competition between degradation and chain extension during processing of reclaimed poly(ethylene terephthalate). Polymer Degradation and Stability 61, 417–420.

Paci, M., La Mantia, F.P., 1999. Influence of small amounts of polyvinylchloride on the recycling of polyethylene terephthalate. Polymer Degradation and Stability 63, 11–14.

Pan, W., Bai, Z., Su, T., Wang, Z., 2018. Enzymatic degradation of poly(butylene succinate) with different molecular weights by cutinase. International Journal of Biological Macromolecules 111, 1040–1046. Available at https://doi.org/10.1016/j.ijbiomac.2018.01.107.

Pathak, V.M., Navneet, 2017. Review on the current status of polymer degradation: a microbial approach. Bioresources and Bioprocessing 4 (1). Available at https://doi.org/10.1186/s40643-017-0145-9, https://bioresourcesbioprocessing.springeropen.com.

Pavia, D.L., Lampman, G.M., Kriz, G.S., 1988. Introduction to Organic Laboratory Techniques, third ed. Saunders, Fort Worth, TX.

Pereira, E.B., De Castro, H.F., De Moraes, F.F., Zanin, G.M., 2001. Kinetic studies of lipase from Candida rugosa. Applied Biochemistry and Biotechnology 91, 739.

Phua, S.K., Castillo, E., Anderson, J.M., Hiltner, A., 1987. Biodegradation of a polyurethane in vitro. Journal of Biomedical Materials Research 21 (2), 231–246. Available at https://doi.org/10.1002/jbm.820210207.

Pitt, C.G., 1992. Non-microbial degradation of polyesters: mechanisms and modifications. Royal Society of Chemistry 7 (12), 180.

Pometto, A.L., Lee, B.T.,, Johnson, K.E., 1992. Production of an extracellular polyethylene-degrading enzyme(s) by Streptomyces species. Applied and Environmental Microbiology 58, 731–733.

Price, D., Horrocks, A.R., 2013. Combustion processes of textile fibres. Handbook of Fire Resistant Textiles. Elsevier Ltd, United Kingdom, pp. 3–25. Available at http://www.sciencedirect.com/science/book/9780857091239, https://doi.org/10.1533/9780857098931.1.3.

Restrepo-Flórez, J.M., Bassi, A., Thompson, M.R., 2014. Microbial degradation and deterioration of polyethylene—a review. International Biodeterioration & Biodegradation 88, 83–90.

Ribitsch, D., Heumann, S., Trotscha, E., Herrero Acero, E., Greimel, K., Leber, R., et al., 2011. Hydrolysis of polyethyleneterephthalate by p-nitrobenzylesterase from Bacillus subtilis. Biotechnology Progress 27 (4), 951–960. Available at https://doi.org/10.1002/btpr.610.

Rizzarelli, P., Carroccio, S., 2009. Thermo-oxidative processes in biodegradable polybutylene succinate. Polymer Degradation and Stability 94, 1825–1838.

Ronqvist, Å.M., Xie, W., Lu, W., Gross, R.A., 2009. Cutinase-catalyzed hydrolysis of poly (ethyleneterephthalate). Macromolecules 42, 5128–5138. Available at https://doi.org/10.1021/ma9005318.

Rowe, L., Howard, G.T., 2002. Growth of Bacillus subtilis on polyurethane and the purification and characterization of a polyurethanase-lipase enzyme. International Biodeterioration and Biodegradation 50 (1), 33–40. Available at https://doi.org/10.1016/S0964-8305(02)00047-1.

Ruiz, C., Howard, G.T., 1999. Nucleotide sequencing of a polyurethanase gene (pulA) from Pseudomonas fluorescens. International Biodeterioration and Biodegradation 44 (2–3), 127–131. Available at https://doi.org/10.1016/S0964-8305(99)00074-8, www.elsevier.com/inca/publications/store/4/0/5/8/9/9.

Russell, J.R., Huang, J., Anand, P., Kucera, K., Sandoval, A.G., Dantzler, K.W., et al., 2011. Biodegradation of polyester polyurethane by endophytic fungi. Applied and Environmental Microbiology 77 (17), 6076–6084. Available at https://doi.org/10.1128/aem.00521-11.

Ru, J., Huo, Y., Yang, Y., 2020. Microbial degradation and valorization of plastic wastes. Frontiers in Microbiology 11, 442–452. Available at https://doi.org/10.3389/fmicb.2020.00442.

Santo, M., Weitsman, R., Sivan, A., 2013. The role of the copper binding enzyme laccase in the biodegradation of polyethylene by the actinomycete *Rhodococcus ruber*.

International Biodeterioration & Biodegradation 84, 204–210. Available at https://doi.org/10.1016/j.ibiod.2012.03.001.

Seo, J.S., Keum, Y.S., Li, Q.X., 2009. Bacterial degradation of aromatic compounds. International Journal of Environmental Research 6, 278–279.

Shah, A.A., Hasan, F., Hameed, A., Ahmed, S., 2008. Biological degradation of plastics: a comprehensive review. Biotechnology Advances 26 (3), 246–265. Available at https://doi.org/10.1016/j.biotechadv.2007.12.005.

Shah, A.A., Eguchi, T., Mayumi, D., Kato, S., Shintani, N., Kamini, N.R., et al., 2013. Degradation of aliphatic and aliphatic-aromatic co-polyesters by depolymerases from Roseateles depolymerans strain TB-87 and analysis of degradation products by LC-MS. Polymer Degradation and Stability 98 (12), 2722–2729. Available at https://doi.org/10.1016/j.polymdegradstab.2013.10.003.

Sharma, P.K., Mohanan, N., Sidhu, R., Levin, D.B., 2019. Colonization and degradation of polyhydroxyalkanoates by lipase-producing bacteria. Canadian Journal of Microbiology 65 (6), 461–475. Available at https://doi.org/10.1139/cjm-2019-0042, https://www.nrcresearchpress.com/journal/cjm.

Shimao, M., 2001. Biodegradation of plastics. Current Opinion in Biotechnology 12 (3), 242–247. Available at https://doi.org/10.1016/s0958-1669(00)00206-8.

Shi, K., Su, T., Wang, Z., 2019. Comparison of poly (butylene succinate) biodegradation by *Fusarium solani* cutinase and Candida antarctica lipase. Polymer Degradation and Stability 164, 55–60.

Sivan, A., 2011. New perspectives in plastic biodegradation. Current Opinion in Biotechnology 22, 422–426.

Son, H.F., Cho, I.J., Joo, S., Seo, H., Sagong, H.Y., Choi, S.Y., et al., 2019. Rational protein engineering of thermo-stable PETase from Ideonella sakaiensis for highly efficient PET degradation. ACS Catalysis 9 (4), 3519–3526. Available at https://doi.org/10.1021/acscatal.9b00568, http://pubs.acs.org/page/accacs/about.html.

Srikanth, M., Sandeep, T.S.R.S., Sucharitha, K., Godi, S., 2022. Biodegradation of plastic polymers by fungi: a brief review. Bioresources and Bioprocessing 9 (1), 42.

Sriyapai, P., Chansiri, K., Sriyapai, T., 2018. Isolation and characterization of polyester-based plastics-degrading bacteria from compost soils. Microbiology (Reading, England) 87, 290–300.

Sriyapai, P., Sriyapai, T., Sukrakanchana, L., 2019. Optimization of polybutylene succinate (PBS)-degrading enzyme production from *Saccharothrix sp. APL5*. Burapha Science Journal 24, 1160–1176.

Stepczyńska, M., Rytlewski, P., 2018. Enzymatic degradation of flax-fibers reinforced polylactide. International Biodeterioration & Biodegradation 126, 160–166.

Tahir, L., Ali, M.I., Zia, M., Atiq, N., Hasan, F., Ahmed, S., 2013. Production and characterization of esterase in Lantinus tigrinus for degradation of polystyrene. Polish Journal of Microbiology 62 (1), 101–108. Available at https://doi.org/10.33073/pjm-2013-015, http://www.pjm.microbiology.pl/full/vol6212013.pdf.

Tan, Y., Henehan, G.T., Kinsella, G.K., Ryan, B.J., 2021. An extracellular lipase from Amycolatopsis mediterannei is a cutinase with plastic degrading activity. Computational and Structural Biotechnology Journal 19, 869–879. Available at https://doi.org/10.1016/j.csbj.2021.01.019, www.csbj.org.

Thirunavukarasu, K., Edwinoliver, N.G., Durai Anbarasan, S., Gowthaman, M.K., Iefuji, H., Kamini, N.R., 2008. Removal of triglyceride soil from fabrics by a novel lipase from

Cryptococcus sp. S-2. Process Biochemistry 43 (7), 701–706. Available at https://doi.org/10.1016/j.procbio.2008.02.011.

Thirunavukarasu, K., Purushothaman, S., Sridevi, J., Aarthy, M., Gowthaman, M.K., Nakajima-Kambe, T., et al., 2016. Degradation of poly(butylene succinate) and poly (butylene succinate-co-butylene adipate) by a lipase from yeast Cryptococcus sp. grown on agro-industrial residues. International Biodeterioration and Biodegradation 110, 99–107. Available at https://doi.org/10.1016/j.ibiod.2016.03.005, www.elsevier.com/inca/publications/store/4/0/5/8/9/9.

Tokiwa, Y., Suzuki, T., 1977a. Hydrolysis of polyesters by lipases. Nature 270, 76–78.

Tokiwa, Y., Suzuki, T., 1977b. Purification of polyethylene adipate-degrading enzyme produced by *Penicillium sp.* strain 14-3. Agricultural and Biological Chemistry 41, 265–274.

Tokiwa, Y., Calabia, B.P., 2004. Degradation of microbial polyesters. Biotechnology Letters 26, 1181–1189.

Tokiwa, Y., Calabia, B.P., 2009. Biodegradability of plastics. International Journal of Molecular Sciences 10, 3722–3742.

Tokiwa, Y., Ando, T., Suzuki, T., 1976. Degradation of polycaprolactone by a fungus. Journal of Fermentation Technology 54, 603–608.

Tokiwa, Y., Calabia, B.P., Ugwu, C.U., Aiba, S., 2009. Biodegradability of plastics. International Journal of Molecular Sciences 10 (9), 3722–3742.

Trevino, A.L., Gutiérrez-Sánchez, G., Rodríguez-Herrera, R., Aguilar, C.N., 2012. Microbial enzymes involved in polyurethane biodegradation: a review. Journal of Polymers and the Environment 20, 258–265.

Tsushima, S., Matsushita, Y., 2010. Technical Report on the PCR-DGGE Analysis of Bacterial and Fungal Soil Communities. National Institute for Agro-Environmental Sciences, Tsukuba. ver. 3.3.

Uchida, H., Nakajima-Kambe, T., Shigeno-Akutsu, Y., Nomura, N., Tokiwa, Y., Nakahara, T., 2000. Properties of a bacterium which degrades solid poly(tetramethylene succinate)-co-adipate, a biodegradable plastic. FEMS Microbiology Letters 189 (1), 25–29. Available at https://doi.org/10.1111/j.1574-6968.2000.tb09201.x.

Umare, S.S., Chandure, A.S., Pandey, R.A., 2007. Synthesis, characterization and biodegradable studies of 1, 3-propanediol based polyesters. Polymer Degradation and Stability 92, 464–479.

Urbanek, A.K., Miron´czuk, A.M., García-Martín, A., Saborido, A., Mata, I., Arroyo, M., 2020. Biochemical properties and biotechnological applications of microbial enzymes involved in the degradation of polyester-type plastics. Biochimica et Biophysica Acta (BBA) - Proteins and Proteomics 1868, 140315.

van Gemeren, I.A., Beijersbergen, A., van den Hondel, C.A.M.J.J., Verrips, C.T., 1998. Expression and secretion of defined cutinase variants by Aspergillus awamori. Applied and Environmental Microbiology 64 (8), 2794–2799. Available at https://doi.org/10.1128/aem.64.8.2794-2799.1998.

Vega, R.E., Main, T., Howard, G.T., 1999. Cloning and expression in *Escherichia coli* of apolyurethane-degrading enzyme from *Pseudomonas fluorescens*. International Biodeterioration & Biodegradation 43, 49–55.

Vertommen, M.A.M.E., Nierstrasz, V.A., van der Veer, M., Warmoeskerken, M.M.C.G., 2005. Enzymatic surface modification of poly(ethylene terephthalate). Journal of Biotechnology 120 (4), 376–386. Available at https://doi.org/10.1016/j.jbiotec.2005.06.015.

Villain, F., Coudane, J., Vert, M., 1995. Thermal degradation of polyethylene terephthalate: study of polymer stabilization. Polymer Degradation and Stability 49, 393–397.

Vona, I.A., Costanza, J.R., Cantor, H.A., Roberts, W.J., 1965. Manufacture of Plastics, vol. 1. Wiley, pp. 141–142

Waring, R.H., Harris, R.M., Mitchell, S.C., 2018. Plastic contamination of the food chain: a threat to human health? Maturitas 115, 64–68.

Webb, H.K., Arnott, J., Crawford, R.J., Ivanova, E.P., 2013. Plastic degradation and its environmental implications with special reference to poly(ethylene terephthalate). Polymers 5 (1), 1–18. Available at https://doi.org/10.3390/polym5010001Australia, http://www.mdpi.com/2073-4360/5/1/1/pdf?y=1.

Wei, R., Zimmermann, W., 2017. Microbial enzymes for the recycling of recalcitrant petroleum-based plastics: how far are we? Microbial Biotechnology 10 (6), 1308–1322. Available at https://doi.org/10.1111/1751-7915.12710.

Wilkes, R.A., Aristilde, L., 2017. Degradation and metabolism of synthetic plastics and associated products by Pseudomonas sp.: capabilities and challenges. Journal of Applied Microbiology 123 (3), 582–593. Available at https://doi.org/10.1111/jam.13472.

Williams, D.F., 1981. Enzymic hydrolysis of polylactic acid. Engineering in Medicine 10, 5–7.

Wu, J., Xiao, Y., Yu, H., 2005. Degradation of lignin in pulp mill wastewaters by white-rot fungi on biofilm. Bioresource Technology 96 (12), 1357–1363.

Yamamoto-Tamura, K., Hiradate, S., Watanabe, T., et al., 2015. Contribution of soil esterase to biodegradation of aliphatic polyester agricultural mulch film in cultivated soils. AMB Express 5, 10.

Yamano, N., Kawasaki, N., Ida, S., Nakayama, A., 2019. Biodegradation of polyamide 4 in seawater. Polymer Degradation and Stability 166, 230–236. Available at https://doi.org/10.1016/j.polymdegradstab.2019.05.032.

Yamashita, Y.S., Ueda, H., Koitabashi, M., Kitamoto, H., 2019. Pretreatment with an esterase from the yeast *Pseudozyma antarctica* accelerates biodegradation of plastic mulch film in soil under laboratory conditions. Journal of Bioscience and Bioengineering 127 (1), 93–98.

Yasuhira, K., Uedo, Y., Takeo, M., Kato, D.I., Negoro, S., 2007. Genetic organization of nylon-oligomer-degrading enzymes from alkalophilic bacterium, Agromyces sp. KY5R. Journal of Bioscience and Bioengineering 104 (6), 521–524. Available at https://doi.org/10.1263/jbb.104.521.

Yoon, M.G., Jeon, H.J., Kim, M.N., 2012. Biodegradation of polyethylene by a soil bacterium and AlkB cloned recombinant cell. Journal of Bioremediation & Biodegradation 3, 1–8. Available at https://doi.org/10.4172/2155-6199.1000145.

Zhang, M., Sharaf, F., Chengtao, L., 2022. Screening and characterization of novel lipase producing *Bacillus* species from agricultural soil with high hydrolytic activity against PBAT poly (butylene adipate co terephthalate) co-polyesters. Polymer Bulletin 79, 10053–10076.